Interactions of Cyclic Hydrocarbons with Biological Membranes

SIKKEMA, J; DEBONT, JAM; POOLMAN, B

Published in:
The Journal of Biological Chemistry

Publication date:
1994

THE JOURNAL OF BIOLOGICAL CHEMISTRY
Vol. 269, No. 11, Issue of March 18, pp. 8022-8028, 1994
Printed in U.S.A.

Interactions of Cyclic Hydrocarbons with Biological Membranes*

(Received for publication, June 14, 1993, and in revised form, November 29, 1993)

Jan Sikkema‡§, Jan A. M. de Bont‡, and Bert Poolman¶

From the ‡Division of Industrial Microbiology, Department of Food Science, Wageningen Agricultural University, P. O. Box 8129, 6700 EV Wageningen and the ¶Department of Microbiology, University of Groningen, P. O. Box 14, 9750 AA Haren, The Netherlands

Many cyclic hydrocarbons, *e.g.* aromatics, cycloalkanes, and terpenes, are toxic to microorganisms. The primary site of the toxic action is probably the cytoplasmic membrane, but the mechanism of the toxicity is still poorly understood. The effects of cyclic hydrocarbons were studied in liposomes prepared from *Escherichia coli* phospholipids. The membrane-buffer partition coefficients of the cyclic hydrocarbons revealed that these lipophilic compounds preferentially reside in the membrane. The partition coefficients closely correlated with the partition coefficients of these compounds in a standard octanol-water system. The accumulation of hydrocarbon molecules resulted in swelling of the membrane bilayer, as assessed by the release of fluorescence self-quenching of fluorescent fatty acid and phospholipid analogs. Parallel to the expansion of the membrane, an increase in membrane fluidity was observed. These effects on the integrity of the membrane caused an increased passive flux of protons and carboxyfluorescein. In cytochrome *c* oxidase containing proteoliposomes, both components of the proton motive force, the pH gradient and the electrical potential, were dissipated with increasing concentrations of cyclic hydrocarbons. The dissipating effect was primarily the result of an increased permeability of the membrane for protons (ions). At higher concentrations, cytochrome *c* oxidase was also inactivated. The effective concentrations of the different cyclic hydrocarbons correlated with their partition coefficients between the membrane and aqueous phase. The impairment of microbial activity by the cyclic hydrocarbons most likely results from hydrophobic interaction with the membrane, which affects the functioning of the membrane and membrane-embedded proteins.

Cyclic hydrocarbons, such as aromatics, alicyclics, and terpenes, interact with biological membranes (de Smet *et al.*, 1978; Sikkema *et al.*, 1992; Uribe *et al.*, 1985, 1990). These interactions lead to changes in structure and function of the membranes, which in turn, may impair growth and activity of the cells (Sikkema *et al.*, 1992). The widespread use of cyclic hydrocarbons (*e.g.* fuels, solvents, starting compounds for organic synthesis) and their release in the environment makes knowledge of their metabolism and toxicity of eminent importance. The toxicity of cyclic hydrocarbons has been well noted (Smith, 1993), but knowledge about their mode of interaction with cells and the cause of toxicity is scarce. Uribe and coworkers studied the toxicity of β-pinene (Uribe *et al.*, 1985) and cyclohexane (Uribe *et al.*, 1990) on intact yeast cells and isolated mitochondria. Both compounds exerted their action at the level of the membrane and membrane-embedded enzymes. Recently, we have reported the effects of the aromatic hydrocarbon tetralin on the structure and function of both bacterial and liposomal membranes (Sikkema *et al.*, 1992). Our data showed that tetralin accumulated in the membrane (partition coefficient approximately 1,100), causing "expansion" of the membrane surface area, inhibition of primary ion pumps, and increase in proton permeability. As a result the electrical potential and pH gradient were dissipated, which may have been the primary cause of inhibition of cellular growth. Further experiments with other aromatic and alicyclic hydrocarbons indicated that the observed effects were not specific for tetralin and that a direct relationship can be found between the partitioning of a particular compound in the membrane and its effect on the structural integrity and functional properties of the membrane (this paper). Effects of polar and non-polar compounds on biological membranes have been reported for fatty acids (Rottenberg, 1990), ethanol in yeast (Cartwright *et al.*, 1986; Leão and van Uden, 1984), and anesthetics in erythrocytes (Seeman, 1972). The explanation most given for the observed toxicity of these compounds is disruption of membrane structure by hydrophobic interaction with the lipid bilayer due to their lipophilicity.

In this investigation, the toxic effects of different cyclic hydrocarbons were studied and related to their hydrophobicity and partitioning into the membrane. The results show that effects of cyclic hydrocarbons on structural and functional properties of membranes are closely related to their accumulation in the membrane. The data give a rationale for the frequently observed correlation between the toxicity of lipophilic compounds to microorganisms and the partition coefficients of such compounds in a standard octanol-water system (log P or k_{OW}; Leo *et al.*, 1971).

MATERIALS AND METHODS

Preparation of Liposomes—Escherichia coli phospholipids, obtained from Sigma, were washed with acetone/ether (Kagawa and Racker, 1971). The commercially obtained *E. coli* lipids contained phosphatidylethanolamine (72 mol %), lyso-phosphatidylethanolamine (5.2 mol %), and cardiolipin (20.5 mol %) (In 't Veld *et al.*, 1992). Lipids dissolved in $CHCl_3$/MeOH (9:1, v/v) were mixed in appropriate quantities and dried under a stream of N_2 gas. Traces of solvent were then removed under vacuum for 1 h. Dried lipid was suspended in 50 mM potassium phosphate (pH 7.0) at a concentration of 20 mg lipid/ml and dispersed by ultrasonic irradiation using a bath sonicator (Sonicor, Sonicor Instruments, New York). Single membrane liposomes (Chapman, 1984; Elferink *et al.*, 1992) were obtained by sonication (probe type sonicator, MSE, West Sussex, United Kingdom) for 300 s at maximal amplitude, using intervals of 15-s sonication and 45-s rest, at 4 °C under a constant stream of N_2 gas.

Partitioning of Lipophilic Compounds—Partitioning of lipophilic compounds over membrane and buffer phases was determined in a *E.*

* This work was supported by the Dutch Programme Committee on Industrial Biotechnology. The costs of publication of this article were defrayed in part by the payment of page charges. This article must therefore be hereby marked "*advertisement*" in accordance with 18 U.S.C. Section 1734 solely to indicate this fact.

§ To whom correspondence should be addressed: Snow Brand European Research Laboratories BV, Zernikepark 6, 9747 AN, Groningen, The Netherlands. Fax: +31-50-745766.

coli phospholipid liposome/potassium phosphate buffer system (De Young and Dill, 1988; Katz and Diamond, 1974). Increasing amounts of the radiolabeled compounds were added to 50 mM potassium phosphate (pH 7.0) containing liposomes (5.0 mg of phospholipid/ml; final volume 0.5 ml). After equilibration (30 min), the liposomes were spun down in an Airfuge (Beckman Instruments) for 30 min at 135,000 × *g*. By this method all liposomes were pelleted, as was assessed by phosphate analyses of control incubations performed in MOPS[1] buffer (50 mM, pH 7) (Rouser *et al.*, 1970). The supernatant was removed with a Pasteur pipette, and two portions of 100 µl were pipetted in a scintillation vial. The pellet was resuspended in scintillation fluid. Both the pellet and the supernatant fractions were analyzed radiometrically in a scintillation counter. The results presented are the mean and the standard deviation of six independent measurements. Control experiments without liposomes were performed in parallel to account for losses of solvent due to possible evaporation and/or attachment to tubes and pipettes. The internal volume of the liposomes (3 µl/mg phospholipid) was taken into account. Partition coefficients of the non-radioactive lipophilic compounds carvone and tetralin were determined by gas chromatography, as described previously (Sikkema *et al.*, 1992).

Membrane Expansion and Extraction of Phospholipids—The expansion of liposomal membranes and extraction of phospholipids from the liposomes due to the addition of lipophilic compounds was monitored in liposomes labeled with the fluorescent fatty acid, octadecyl rhodamine-β-chloride (R_{18}; Molecular Probes Inc., Junction City, OR) or the fluorescent phospholipid analog, *N*-(lissamine rhodamine-β-sulfonyl)phosphatidylethanolamine (*N*-Rh-PE; Avanti Polar Lipids Inc., Alabaster, AL). The method is based on the relief of fluorescence self-quenching (Hoekstra *et al.*, 1984) of rhodamine-β-chloride as a result of expansion of the membrane and/or extraction of the probe from the membrane. The fatty acid probe was incorporated into liposomal membranes at a concentration of 4 mol % phospholipid phosphorous. Maximum rhodamine fluorescence was determined upon the addition of 1% (v/v) Triton X-100. Fluorescent changes were measured in a spectrofluorometer (Perkin-Elmer Cetus) using the excitation-emission pair 560 and 590 nm. In order to discriminate between fluorescence increases due to expansion of the membrane and extraction of membrane constituents, incubation mixtures with different concentrations of lipophilic compounds were centrifuged at 135,000 × *g* (Beckman Airfuge, 30 min). Subsequently, the fluorescence of the supernatant was determined relative to the supernatant of an incubation without hydrocarbon added. Additionally, supernatants of incubations containing *E. coli* phospholipid liposomes in MOPS (50 mM, pH 7.0) and varying amounts of cyclic hydrocarbons were assayed for released phospholipids by phosphate analysis (Rouser *et al.*, 1970).

Membrane Fluidity Measurements/Fluorescence Polarization Measurements—DPH (1,6-diphenyl-1,3,5-hexatriene) and TMA-DPH (1-[4-(trimethylamino)phenyl]-6-phenylhexa-1,3,5-triene) steady-state polarization measurements were carried out as described (In 't Veld *et al.*, 1991). Membrane fluidity is used as a qualitative measure and is defined as the inverse of microviscosity. Microviscosity can be deduced from the steady-state fluorescence polarization of (TMA-)DPH probes (Shinitzky and Barenholz, 1978).

The degree of fluorescence polarization was calculated from Equation 1 (Lentz, 1989; Shinitzky and Barenholz, 1978):

$$r_{ss} = \frac{I_{\parallel} - I_{\perp}}{I_{\parallel} + 2I_{\perp}} \qquad \text{(Eq. 1)}$$

r_{ss}, steady-state fluorescence polarization; $I_{\parallel}$, fluorescence intensity at 430 nm, measured parallel to the emitted light; $I_{\perp}$: fluorescence intensity at 430 nm, measured perpendicular to the emitted light.

Reconstitution of Cytochrome c Oxidase into Proteoliposomes—Aceton-ether-washed *E. coli* lipid (40 mg) and *n*-octyl-β-D-glucopyranoside (18 mg) in 2 ml of 50 mM potassium phosphate (pH 7.0) was cosonicated until clarity under a constant stream of N_2 gas at 4 °C using a probe sonicator. Cytochrome *c* oxidase (9 nmol of heme *a* was added, and the suspension was dialyzed at 4 °C for 4 h against a 500-fold volume of 50 mM potassium phosphate (pH 7.0). Dialysis was repeated for another 4 h and continued overnight at 4 °C (Hinkle *et al.*, 1972).

Internal pH of Cytochrome c Oxidase Containing Proteoliposomes—Internal pH changes were measured by following the fluorescence of entrapped pyranine (Eastman Kodak Co.) (Clement and Gould, 1981). To incorporate pyranine into proteoliposomes (20 mg phospholipid/ml), 100 nmol of pyranine was added to 0.5 ml of proteoliposomes and rapidly mixed. The suspensions were rapidly frozen in liquid nitrogen and subsequently thawed slowly (approximately 30 min) at room temperature. The suspension was sonicated for 8 s using a probe type sonicator at an amplitude of 4. To remove external pyranine, the proteoliposomes were washed in 10 ml of 50 mM potassium phosphate (pH 7.0) and centrifuged for 45 min at 280,000 × *g* in a Beckman type Ti-75 rotor at 4 °C. Fluorescent changes were measured at excitation and emission wavelengths of 460 and 508 nm, respectively. Calibration was performed by titration with acid or base upon addition of nigericin to a final concentration of 20 nM. At pH 7.0, the ΔpH generated by the cytochrome *c* oxidase-containing proteoliposomes was 0.8–0.9.

Electrical Potential across Membranes of Proteoliposomes—The transmembrane electrical potential ($\Delta\psi$) of cytochrome *c* oxidase containing liposomes was determined by monitoring the distribution of tetraphenylphosphonium (TPP^+) across the membrane with a TPP^+-sensitive electrode as previously described (Lolkema *et al.*, 1982).

Proton Fluxes through Liposomal Membranes—$\Delta\psi$-Induced proton fluxes were estimated in liposomes in the presence of varying concentrations of a particular hydrocarbon compound in a well stirred thermostated 2-ml cuvette, using phenol red (20 µg/ml, final concentration) as external pH indicator. Absorbance changes ($A_{560} - A_{610}$) were converted into H^+ fluxes by calibrating with known amounts of oxalic acid or KOH (de Vrij *et al.*, 1988). Valinomycin-induced potassium diffusion potentials were imposed across the liposomal membrane by 100-fold dilution of the liposomes (20 mg of phospholipid/ml) into the same medium, in which sodium ions were substituted for potassium ions and supplemented with phenol red (20 µg/ml). Generation of the electrical potential was initiated by adding valinomycin (2 µM, final concentration).

Determination of Cytochrome c Oxidase Activity—Cytochrome *c* oxidase activity was measured spectrophotometrically by monitoring the decrease in the absorbance of the alpha peak of reduced cytochrome *c*, using an extinction coefficient (reduced minus oxidized) of $\epsilon_{550-540}$ = 19.5 mM^{-1} cm^{-1} (de Vrij *et al.*, 1988).

Measurement of Carboxyfluorescein Efflux—Release of carboxyfluorescein (CF) from liposomes, resulting in relief of its fluorescence self-quenching, was determined in a Perkin Elmer spectrofluorometer equipped with a thermostated cell holder, using the excitation-emission pair 430 and 520 nm, respectively. Encapsulation of 5,6-carboxyfluorescein (CF, Eastman Kodak Chemical Co.) was achieved by preparing liposomes in 50 mM CF following the protocol for the formation of pyranine containing liposomes (see above).

Chemicals—All hydrocarbons used were of the highest available commercial grade. Radiochemicals were obtained from the Radiochemical Centre, Amersham, United Kingdom ([^{14}C]phenylalanine, [^{14}C]toluenesulfonic acid, [^{14}C]benzoic acid, [^{14}C]4-chlorobenzoic acid) and Sigma ([^{14}C]toluene, [^{14}C]naphthalene, and [^{14}C]phenanthrene. [^{3}H]TPP^+, used for the determination of the partition coefficient, was obtained from Amersham.

Addition of Cyclic Hydrocarbons—The hydrocarbons were prepared as solutions in *N*-dimethylformamide (DMF). In all cases, the amount of DMF was 2% (v/v) of the total volume. In this concentration DMF had no effect on any parameter studied except for the binding of TPP^+ to membranes (binding of TPP^+ was less in the presence of DMF); $\Delta\psi$ values were corrected accordingly (Lolkema *et al.*, 1982).

RESULTS

Partitioning of Lipophilic Compounds—In order to gain insight in the effects of lipophilic compounds, at subsaturating concentrations in the aqueous phase, on biological membranes it is essential to know the partitioning behavior of such compounds in a membrane-buffer system. As a model system for a biological membrane, liposomes were used prepared from *E. coli* phospholipids.

Membrane-buffer partition coefficients in the *E. coli* phospholipid liposomes/potassium phosphate buffer were determined for compounds varying in hydrophobicity/lipophilicity, *i.e.* phenylalanine, benzoic acid, 4-chlorobenzoic acid, 4-toluenesulfonic acid, TPP^+, carvone, toluene, naphthalene, tetralin, and phenanthrene. The choice of most of these molecules stems from their availability in radioactive form. As a measure for the hydrophobicity of the compounds, the octanol-water partition

[1] The abbreviations used are: MOPS, 4-morpholinepropanesulfonic acid; DPH, 1,6-diphenyl-1,3,5-hexatriene; TMA-DPH, 1-[4-(trimethylamino)phenyl]-6-phenylhexa-1,3,5-triene; CF, carboxyfluorescein; DMF, *N*-dimethylformamide; TMPD; *N,N,N',N'*-tetramethyl-*p*-phenylenediamine.

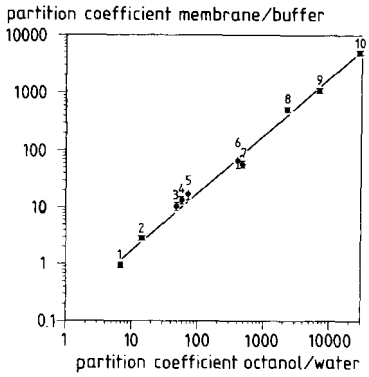

FIG. 1. **Relationship between the partition coefficients in an *E. coli* phospholipid membrane-potassium phosphate buffer (pH 7.0; 50 mM) system and the partition coefficients in the standard *n*-octanol-water system of 4-toluenesulfonic acid (*1*), 4-chlorobenzoic acid (*2*), benzoic acid (*3*), phenylalanine (*4*), TPP^+ (*5*), carvone (*6*), toluene (*7*), naphthalene (*8*), tetralin (*9*), and phenanthrene (*10*).** The experimental points represent the mean and standard deviation of six independent measurements.

coefficients were taken (Leo *et al.*, 1971). The distribution of the lipophilic compounds over the aqueous and the lipid phase was determined at several solvent to lipid ratios. When the measured aqueous concentration was plotted against the lipid to solvent ratio, a saturation curve was obtained (Sikkema *et al.*, 1992). The partition coefficient was calculated, from the linear part of this curve (below maximum aqueous solubility). The membrane-buffer partition coefficients were plotted as a function of the octanol-water partition coefficients (Fig. 1). Despite differences in structural features of the molecules, a good correlation between the partitioning in a membrane-buffer system and a standard octanol-water system was observed. The correlation line for lipophilic compounds with logP values between approximately 1 and 4.5, is described by Equation 2:

$$\log P_{M/B} = 0.97 \times \log P_{O/W} - 0.64 \qquad \text{(Eq. 2)}$$

The correlation coefficient for the four aromatic hydrocarbons toluene, naphthalene, tetralin, and phenanthrene is 0.9967. With this equation, the membrane-buffer partition coefficients of 20 cyclic hydrocarbons were calculated from their octanol-water partition coefficients. In Table I the membrane-buffer partition coefficients of these cyclic hydrocarbons together with other physical and chemical data of these compounds are given.

Expansion of the Membrane—Due to the accumulation of lipophilic compounds in the lipid bilayer, changes in the membrane structure and even swelling of the membrane can be expected. The effect of accumulation in the membrane surface area was monitored by using liposomes prepared from *E. coli* phospholipids that were labeled with R_{18} or *N*-Rh-PE. The rationale of this method is that expansion of the membrane leads to dilution of the probe in the membrane which can be measured as a relief in fluorescence self-quenching. Since the fluorescence signal is related to the lipid concentration (Hoekstra *et al.*, 1984), a change in fluorescence will be proportional to a change in surface area. An increase in fluorescence could, however, also be due to extraction of the fluorescent probe from the membrane by the hydrocarbon. Ultracentrifugation of liposomes equilibrated with varying amounts of toluene, cyclohexane, and tetralin showed that at the most 16.3, 11, and 9.4% of the fluorescence increase with 150 μmol of toluene, 15 μmol of cyclohexane, and 5 μmol of tetralin/mg phospholipid, respectively, could be attributed to probe extraction from the membrane. In addition, supernatants of incubations containing liposomes and varying concentrations of hydrocarbon were checked for the presence of free phospholipids. The highest concentrations of each hydrocarbon applied in the experiments with R_{18}-labeled liposomes (see Fig. 2) did not result in extraction of more than 10% of the phospholipid content. The data for the different compounds were: decalin, 8.6% of total phospholipid phosphate solubilized at 3 μmol/mg PL; anthracene, 8.4% at 1 μmol/mg PL; biphenyl, 9.0% at 2.5 μmol/mg PL; α-pinene, 9.3% at 2.5 μmol/mg PL; tetralin, 6.2% at 5 μmol/mg PL; naphthalene, 7.8% at 6 μmol/mg PL; cyclohexane, 9.9% at 15 μmol/mg PL; *o*-xylene, 9.6% at 60 μmol/mg PL; ethylbenzene, 8.7% at 70 μmol/mg PL; toluene, 9.4% at 150 μmol/mg PL; benzene, 9.1% at 250 μmol/mg PL. At higher concentrations solubilization of the liposomes did occur, which was not only detected by a rapid increase of free phospholipids in the supernatant but also by the increase of turbidity of the suspension in the cuvette. In a set of control experiments it was shown that the hydrocarbon solvent had no direct effect on fluorescence intensity, which could have occurred as a result of modification of the microenvironment of the probe. In these experiments the same concentrations of solvents (Fig. 2) were mixed with liposomes labeled with non-self-quenching concentrations of the fluorescent probes. Taken together, these results indicate that the observed increase in R_{18} fluorescence was primarily due to swelling of the membrane. Different solvents exhibit different concentration dependencies and extents of apparent membrane expansion (Fig. 2). For instance, in the presence of decalin the increase in rhodamine fluorescence not only occurred at a much lower concentration than with benzene, but the extent of fluorescence increase was also higher. The difference in effective concentration at which the rhodamine fluorescence increased parallels the change in hydrophobicity of the compounds and the partitioning into the membrane. The differences in the extent of the fluorescence increase could be due to differences in maximum solubility of the hydrocarbon in the membrane but may also reflect differences in location in the membrane. Results similar to those presented in Fig. 2 were obtained with *N*-Rh-PE-labeled liposomes (data not shown).

Changes in Membrane Fluidity as a Result of Interaction with Hydrocarbons—The fluidity of a membrane bilayer can be assessed by determining the fluorescence polarization of DPH or TMA-DPH. Although the precise location of DPH in the membrane is still not clear, this probe most likely resides near the center of the bilayer (Lentz, 1989). Less ambiguities exist about the location of TMA-DPH since its hydrophilic group anchors the molecule at the headgroup region of the bilayer thereby aligning the DPH moiety with the phospholipid acyl chains. All hydrocarbons except biphenyl decreased the polarization of DPH whereas TMA-DPH polarization was not significantly affected (Fig. 3). The different locations in the membrane of DPH and TMA-DPH and the different effects of tetralin, cyclohexane, naphthalene, and toluene on the fluorescence polarization of DPH and TMA-DPH suggest that the hydrocarbons perturb the bilayer structure primarily by accumulating into the interior rather than into the peripheral regions of the membrane.

Effects of Hydrocarbons on the Proton Motive Force—The accumulation of hydrocarbons in the lipid bilayer, and the consequent change in membrane-structure due to membrane-expansion, change in membrane fluidity, and/or disruption of lipid-protein interactions could have a strong effect on the functioning of the membrane as a selective barrier for ions and hydrophilic molecules. The permeability for protons and other ions is especially of importance since ion leaks directly affect the energy transducing properties of the membrane. To analyze the effect of hydrocarbons on the generation of the transmembrane pH gradient (ΔpH) and electrical potential ($\Delta\psi$) in artificial membranes, beef heart mitochondrial cytochrome *c* oxi-

TABLE I
Physical data of the cyclic hydrocarbons used in this study

Compound	Formula	M_r[a]	Solubility[b]	logP	$P_{O/W}$	$P_{M/B}$exp	$P_{M/B}$calc[b,c]
			mmol/l, 25 °C				
Benzene	C_6H_6	78.11	22.9	2.13[b]	135		27
Cyclohexane	C_6H_{12}	84.16	0.683	3.44[b,d]	2,754		498
Toluene	C_7H_8	92.14	6.28	2.69[b]	490	59 ± 8.5	93
Ethylbenzene	C_8H_{10}	106.17	1.27	3.15[b]	1,413		260
o-Xylene	C_8H_{10}	106.17	2.02	3.12[b]	1,318		243
Naphthalene	$C_{10}H_8$	128.17	0.797	3.37[b]	2,344	527 ± 38	426
Tetralin	$C_{10}H_{12}$	132.21	0.125[e]	3.86[d]	7,244	1,100 ± 56	1,271
o-di-Ethylbenzene	$C_{10}H_{14}$	134.22	—[h]	4.10[f]	12,590		2,173
α-Pinene	$C_{10}H_{16}$	136.24	—	4.46[f]	28,840		4,855
β-Pinene	$C_{10}H_{16}$	136.24	—	4.46[f]	28,840		4,855
γ-Terpinene	$C_{10}H_{16}$	136.24	—	4.46[f]	28,840		4,855
Limonene	$C_{10}H_{16}$	136.24	0.101[i]	4.46[f]	28,840		4,855
Decalin	$C_{10}H_{18}$	138.25	—	4.83[f]	67,608		11,094
Biphenyl	$C_{12}H_{10}$	154.21	0.126	4.04[b]	10,965		1,900
Anthracene	$C_{14}H_{10}$	178.23	0.040	4.45[g]	28,184		4,748
Phenanthrene	$C_{14}H_{10}$	178.23	0.025	4.46[g]	28,840	4,937 ± 86	4,855

[a] Data obtained from Handbook of Chemistry and Physics.
[b] Eastcott *et al.*, 1988.
[c] Calculated from $P_{O/W}$ data found in literature and applied to Equation 2.
[d] Rekker, 1977.
[e] Sikkema and de Bont, 1991.
[f] Calculated via fragmental constants (Rekker and de Kort, 1979).
[g] Tomlinson and Hafkenscheid, 1986.
[h] Data not available.
[i] Riddick *et al.*, 1986.

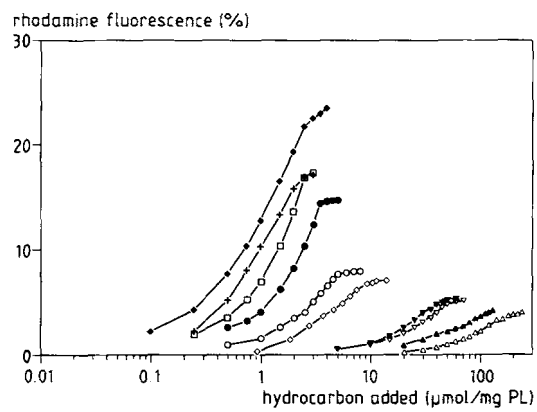

FIG. 2. **Effect of cyclic hydrocarbons on the relief of fluorescence self-quenching of R_{18}-labeled liposomes.** The reaction mixture contained liposomes (0.2 mg of *E. coli* phospholipid/ml) labeled with R_{18} (4 mol %) in potassium phosphate (50 mM, pH 7.0). The changes in fluorescence were monitored using the excitation-emission pair 560 and 590 nm. The temperature of the solution was kept at 30 °C. △, benzene; ▲, toluene; ▽, ethylbenzene; ▼, *o*-xylene; ◇, cyclohexane; ○, naphthalene; ●, tetralin; □, biphenyl; +, α-pinene; ◆, decalin.

dase was reconstituted into liposomes as proton motive force generating mechanism. At an external pH of 7.0, and in the presence of the electron donor system ascorbate-TMPD-cytochrome *c*, cytochrome *c* oxidase containing proteoliposomes generated a $-Z\Delta pH$ and $\Delta\psi$ of −54 and −60 mV, respectively. The results show that all hydrocarbons tested dissipated the ΔpH (Fig. 4) and that the inhibitory concentrations directly correlated with the partitioning of the compound into the membrane as well as with the increase in rhodamine fluorescence and DPH polarization measurements. The $\Delta\psi$ was found to decrease in a similar way as the ΔpH (data not shown).

Site(s) of Action of Hydrocarbons—The observed decrease in ΔpH and $\Delta\psi$ could be the result of an increase in passive proton or ion fluxes, and/or inhibition of the energy transducing activity of the cytochrome *c* oxidase. Incubation of cytochrome *c* oxidase containing liposomes with different concentrations of benzene, cyclohexane, tetralin, decalin, and biphenyl showed

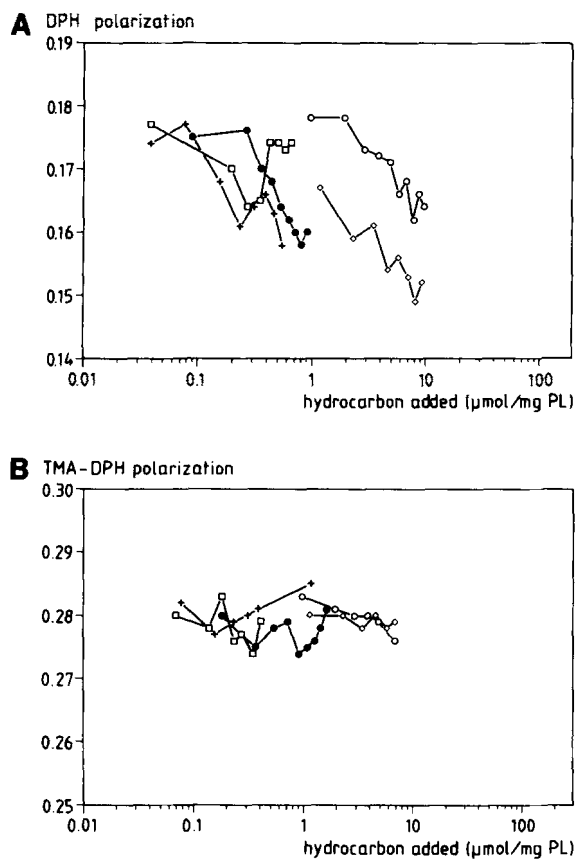

FIG. 3. **Polarization of DPH and TMA-DPH as a result of the interaction of different cyclic hydrocarbons with *E. coli* phospholipid liposomes.** The measurements were performed in a spectrofluorometer at an excitation wavelength of 360 nm; the emission was recorded at 430 nm. The cuvette, thermostated at 30 °C, contained 0.2 mg/ml of either TMA-DPH- or DPH-labeled liposomes (probe to lipid molar ratio in both instances 1–250) in 50 mM potassium phosphate (pH 7.0). ○, naphthalene; ◇, cyclohexane; ●, tetralin; □, biphenyl; +, α-pinene. *A*, effect of hydrocarbons on the polarization of DPH. *B*, effect of hydrocarbons on the polarization of TMA-DPH.

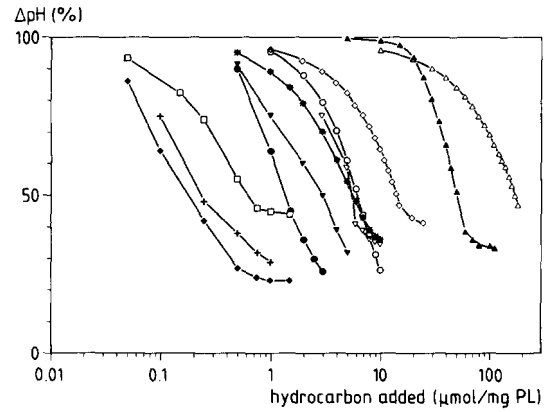

FIG. 4. **Effect of cyclic hydrocarbons on the ΔpH generated by cytochrome *c* oxidase containing proteoliposomes.** Energization of cytochrome *c* oxidase containing proteoliposomes was achieved in the presence of cytochrome *c* (20 μM), TMPD (200 μM), and ascorbate (10 mM). The assay was performed in 50 mM potassium phosphate (pH 7.0), in a cuvette thermostated at 30 °C. △, benzene; ▲, toluene; ▽, ethylbenzene; ▼, *o*-xylene; ○, naphthalene; ◇, cyclohexane; *, *o*-di-ethylbenzene; ●, tetralin; □, biphenyl; +, α-pinene; ◆, decalin.

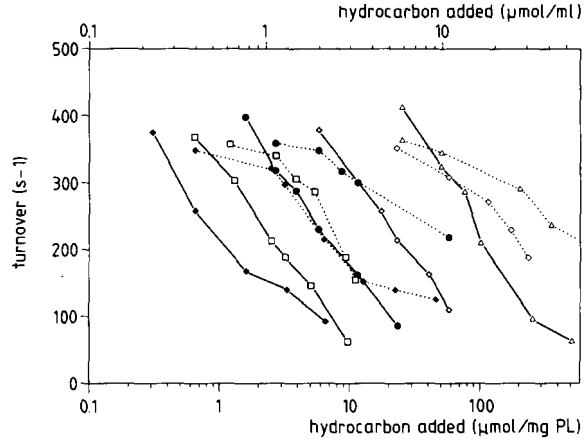

FIG. 5. **Activity of cytochrome *c* oxidase as determined by monitoring the oxidation of reduced cytochrome *c* (A_{550}-A_{540}).** The reaction mixture contained either proteoliposomes (*solid lines*) or Triton X-100- (0.5%, v/v) solubilized proteoliposomes (*dotted lines*), in 50 mM potassium phosphate (pH 7.0); the assay was performed at 30 °C. Δp was decoupled by the addition of valinomycin (2 μM, final concentration), and nigericin (0.1 μM, final concentration). Activity of the cytochrome *c* oxidase is expressed as mole cytochrome *c* per mole enzyme/second. The maximum activity of the cytochrome *c* oxidase in uncoupled liposomes was 425 s^{-1}, and of the enzyme in solubilized liposomes was 370 s^{-1}. △, benzene; ◇, cyclohexane; ●, tetralin; □, biphenyl; ◆, decalin.

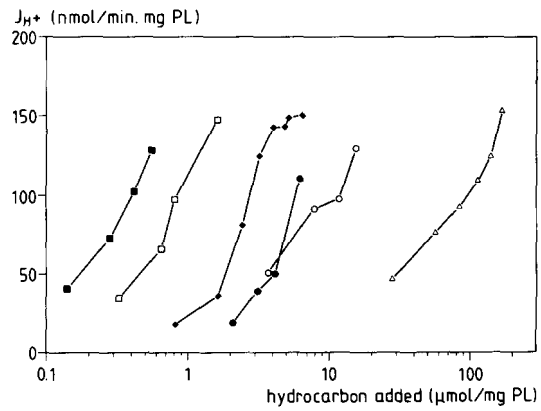

FIG. 6. **The effect of cyclic hydrocarbons on the proton permeability of *E. coli* phospholipid membranes.** Liposomes (0.2 mg of phospholipid/ml) were washed and resuspended in a medium in which sodium ions were substituted for potassium ions and to which phenol red (20 μg/ml) was added. To initiate the potassium diffusion potential, valinomycin (2 μM, final concentration) was added. Subsequently, absorbance changes were measured at A_{560}-A_{610} to determine the external pH changes caused by proton influx as a compensatory effect on the imposed diffusion potential. △, benzene; ○, naphthalene; ●, tetralin; □, biphenyl; ◆, decalin; ■, anthracene.

that indeed inhibition of the enzyme activity occurred. Comparison of the sensitivity of cytochrome *c* oxidase reconstituted in liposomes with the enzyme in Triton X-100 solution indicated that the membrane embedded enzyme was more affected by hydrocarbons (Fig. 5), as could be expected from the accumulation of the molecules in the membrane. Since the enzyme in solution is associated with detergent micelles it is difficult to compare the inhibitory effects on the reconstituted and "free" enzyme quantitatively.

Dissipation of the ΔpH as a result of an increased proton permeability of the membrane was assessed by determining the passive proton influx across the liposomal membrane. Potassium-loaded liposomes were diluted into potassium-free medium in the presence of valinomycin, and the initial rates of H^+ influx in the absence and presence of different amounts of hydrocarbon were determined (Fig. 6). Increasing amounts of hydrocarbon were needed to increase the proton permeability of the membrane going from anthracene, decalin, tetralin, cyclohexane, and toluene to benzene. The concentrations of hydrocarbons that affected the proton permeability were in the same range as those that inhibited cytochrome *c* oxidase.

Permeability of Liposomal Membranes for CF—To assess the effect of cyclic hydrocarbons on the permeability of the membrane for low molecular weight molecules, the efflux of the fluorescent dye CF was examined. In the presence of various hydrocarbons an increased leakage of CF (M_r = 376) was observed that paralleled the increase in permeability of the membrane to protons. The concentration at which leakage of carboxyfluorescein was observed was only slightly higher than the hydrocarbon concentrations needed to increase the proton permeability (data not shown).

DISCUSSION

Due to the hydrophobic character of hydrocarbons, the primary site of their toxicity is the membrane. Hydrocarbons accumulate in the lipid bilayer according to a partition coefficient that is specific for the compound applied. Since partitioning of a compound between a membrane and an aqueous phase is difficult to determine, and may vary with the composition of the membrane, attempts have been made to find a parameter for partitioning. The octanol-water system, which has been applied for many years in anesthesiology and environmental biology (Leo *et al.*, 1971; Verschueren, 1983), proved to be the most suitable model system (Lieb and Stein, 1986). For the *E. coli* phospholipid liposomal membrane-potassium phosphate buffer system, the octanol-water partition coefficient of a wide variety of compounds showed good correlation with the membrane-buffer partition coefficient (Fig. 1). The ratio between these partition coefficients, however, may differ significantly depending on the type of membrane (Antunes-Madeira and Madeira, 1984, 1985, 1986, 1987; Katz and Diamond, 1974; Seeman, 1972). Therefore, each membrane system should be tested before quantitative estimations of the partition coefficients can be made.

The cyclic hydrocarbons were dissolved in DMF in order to increase the dissolution rate of the hydrocarbons. The use of a cosolvent is especially relevant for solid hydrocarbons, such as naphthalene, biphenyl, phenanthrene, and anthracene. By using a cosolvent the distribution of the hydrocarbons in the

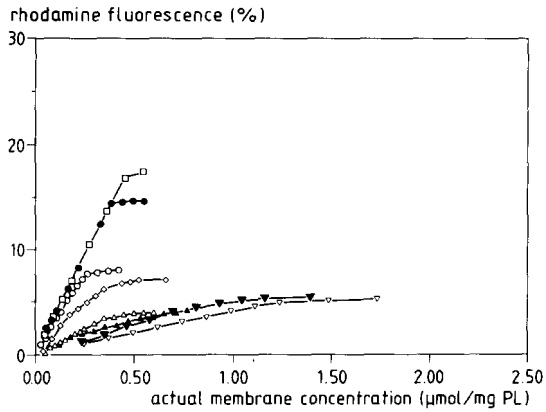

FIG. 7. **Increase in R_{18} fluorescence as a function of the concentration of cyclic hydrocarbons in the membrane as calculated from the membrane-buffer partition coefficient (Table I).** △, benzene; ▲, toluene; ▽, ethylbenzene; ▼, *o*-xylene; ◇, cyclohexane; ○, naphthalene; ●, tetralin; □, biphenyl.

aqueous phase and the membrane phase will come to equilibrium rapidly.

Accumulation of compounds in the membrane may lead to alteration of the membrane structure and function. An important change is the apparent increase in surface area of the membrane, due to swelling of the membrane upon accumulation of lipophilic compounds (Machleidt *et al.*, 1972; Seeman, 1972). The expansion observed with hydrocarbons was more than two times higher than the expansion by alcohols (Seeman *et al.*, 1971). This variation is probably due to differences in the type of hydrophobic interaction and part of the membrane where lipophilic compounds reside (see also below). Differences in the methods applied to determine the increase in surface area were of less importance, since experiments with *n*-alcohols (butanol to decanol) in the *E. coli* phospholipid/R_{18} system gave results that did not significantly differ from the data reported by Seeman and co-workers.[2] The hydrocarbon concentrations that are present in the membrane can be calculated from the estimated membrane-buffer partition coefficients (Table I). When the R_{18} fluorescence data from Fig. 2 are plotted against the membrane concentrations of the hydrocarbons a concentration range at which "swelling" occurs can be seen (Fig. 7). Up to a concentration, in the membrane, of approximately 0.5 μmol/mg phospholipid (±1 hydrocarbon molecule/2 phospholipids) an increase in membrane surface area is observed, after which an apparent maximum is reached. The extent of R_{18} fluorescence increase at an actual membrane concentration higher than 0.5 μmol/mg phospholipid (Fig. 7) was highest for the compounds with the highest $P_{M/B}$, *i.e.* biphenyl and tetralin, naphthalene and cyclohexane were intermediate, whereas *o*-xylene, ethylbenzene, toluene, and benzene were lowest. The cause of this phenomenon is not readily understood although the extent of the R_{18} fluorescence increase parallels the molar volumes of the molecules (Table I).

The increase in membrane fluidity as estimated from DPH polarization measurements (Fig. 3) is already apparent at slightly lower cyclic hydrocarbon concentrations than the increase in membrane surface area (Fig. 2). This is most clear for α-pinene and tetralin, although for cyclohexane and naphthalene this effect can also be seen. No significant effect of the hydrocarbons on the polarization of TMA-DPH was observed. These results suggest that the hydrocarbons partition to the central part of the membrane, which directly affects the polarization of the DPH. In principle one could envisage an effect of hydrocarbons on the distribution of DPH and TMA-DPH in the membrane. However, if for instance DPH would become intercalated among acyl chains, one would expect a decrease in polarization not only with DPH but also with TMA-DPH.

As a result of accumulation of hydrocarbons in the membrane the activity of cytochrome *c* oxidase is lowered and the proton (ion) permeability increases. Both effects act synergically on the magnitude of the ΔpH and $\Delta\psi$ generated by cytochrome *c* oxidase. Since a 50% reduction of cytochrome *c* oxidase activity only causes a small drop in the ΔpH and $\Delta\psi$ (Sikkema *et al.*, 1992), the drop in the components of the proton motive force will primarily be caused by the increased proton permeability. To our knowledge the effects of hydrocarbons on the generation and maintenance of the proton motive force have neither quantitatively nor qualitatively been analyzed so far. Uribe and co-workers (Uribe *et al.*, 1985, 1990) reported results which are in accordance with ours and support the view that an important part of the toxicity of hydrocarbons is exerted by effects on the proton motive force.

The action of general anesthetics on cell functioning, which is similar to the effects observed for cyclic hydrocarbons, is often ascribed to interaction of the anesthetic compounds with the membrane (Overton, 1896; Seeman, 1972). This hypothesis, which ascribes the inhibitory action of anesthetics fully to changes in membrane integrity, is named the lipid theory of anesthesia. The competing theory is the protein-interaction theory, which states that anesthesia is a result of interaction of anesthetic molecules with various enzymes involved in cellular metabolism (Franks and Lieb, 1987). Our studies clearly indicate that the effects of hydrocarbons on the functioning of biological membranes involves both effects on the permeability (protons (ions) but also larger molecules, *e.g.* CF) and the activity of membrane enzymes (cytochrome *c* oxidase). The effects on enzyme activity can be due to altered protein-lipid interactions (hydrogen bonding and others), membrane thickness, fluidity, and/or phospholipid headgroup hydration (Yeagle, 1989). Therefore, it is remarkable that the obvious combination of the lipid theory of anesthesia (Overton, 1896) and the protein-interaction theory advocated by Franks and Lieb ((1987); LaBella, 1981) has not gained more attention so far.

A remarkable outcome of our studies is the observation that the effect of cyclic hydrocarbons on the structural and functional properties of biological membranes ((proteo)liposomes) is directly related to accumulation in these membranes; the effect is independent of the structural features of the molecules. The accumulation of cyclic hydrocarbons in the membranes is proportional to the concentration in the aqueous phase and the membrane-aqueous phase partition coefficient. This latter parameter relates directly to the partitioning of these cyclic hydrocarbons in a standard octanol-water system, which allows predictions to be made about the toxicity of other lipophilic compounds on basis of their logP values. Since bacteria highly differ in their sensitivity toward cyclic hydrocarbons it will be important to establish how the membrane bilayers of these organisms differ and how the $P_{M/B}$ is affected by the phospholipid composition of the membrane. Future studies are aimed at addressing these questions.

Acknowledgments—We thank Prof. Wil Konings for critical reading of the manuscript. Marian Vermuë is gratefully acknowledged for calculating logP values.

[2] J. Sikkema, W. van den Heuvel, and J. A. M. de Bont, unpublished results.

REFERENCES

Antunes-Madeira, M. C., and Madeira, V. M. C. (1984) *Biochim. Biophys. Acta* **778**, 49–56

Antunes-Madeira, M. C., and Madeira, V. M. C. (1985) *Biochim. Biophys. Acta* **820**, 165–172

Antunes-Madeira, M. C., and Madeira, V. M. C. (1986) *Biochim. Biophys. Acta* **861**, 159–164

Antunes-Madeira, M. C., and Madeira, V. M. C. (1987) *Biochim. Biophys. Acta* **901,** 61–66
Antunes-Madeira, M. C., and Madeira, V. M. C. (1989) *Biochim. Biophys. Acta* **982,** 161–166
Cartwright, C. P., Juroszek, J.-R., Beavan, M. J., Ruby, F. M. S., De Morais, S. M. F., and Rose, A. H. (1986) *J. Gen. Microbiol.* **132,** 369–377
Chapman, D. (1984) in *Liposome Technology* (Gregoriadis, G., ed) Vol. 1, pp. 1–18, CRC Press, Boca Raton, FL
Clement, N. R., and Gould, J. M. (1981) *Biochemistry* **20,** 1534–1538
de Smet, M. J., Kingma, J., and Witholt, B. (1978) *Biochim. Biophys. Acta* **506,** 64–80
de Vrij, W., Bulthuis, R. A., and Konings, W. N. (1988) *J. Bacteriol.* **170,** 2359–2366
De Young, L. R., and Dill, K. A. (1988) *Biochemistry* **27,** 5281–5289
Eastcott, L., Shiu, W. Y., and Mackay, D. (1988) *Oil Chem. Poll.* **4,** 191–216
Elferink, M. G. L., de Wit, J. G., Demel, R., Driessen, A. J. M., and Konings, W. N. (1992) *J. Biol. Chem.* **267,** 1375–1381
Franks, N. P., and Lieb, W. R. (1987) *Trends Pharmacol. Sci.* **8,** 169–174
Hinkle, P. C., Kim, J. J., and Racker, E. (1972) *J. Biol. Chem.* **247,** 1338–1339
Hoekstra, D., de Boer, T., Klappe, K., and Wilschut, J. (1984) *Biochemistry* **23,** 5675–5681
In 't Veld, G., Driessen, A. J. M., Op den Kamp, J. A. F., and Konings, W. N. (1991) *Biochim. Biophys. Acta* **1065,** 203–212
In 't Veld, G., de Vrije, T., Driessen, A. J. M., and Konings, W. N. (1992) *Biochim. Biophys. Acta* **1104,** 250–256
Kagawa, Y., and Racker, E. (1971) *J. Biol. Chem.* **235,** 769–775
Katz, Y., and Diamond, J. M. (1974) *J. Membr. Biol.* **17,** 69–86
LaBella, F. S. (1981) *Can. J. Physiol. Pharmacol.* **59,** 432–442
Leão, C., and van Uden, N. (1984) *Biochim. Biophys. Acta* **774,** 43–48
Lentz, B. R. (1989) *Chem. Phys. Lipids* **50,** 171–190
Leo, A., Hansch, C., and Elkins, D. (1971) *Chem. Rev.* **71,** 525–616
Lide, K. (1992) *Handbook of Chemistry and Physics*, CRC Press, Inc. Boca Raton, FL
Lieb, W. R., and Stein, W. D. (1986) in *Transport and Diffusion across Cell Membranes* (Stein, W. D., ed) pp. 69–112, Academic Press Inc., New York
Lolkema, J. S., Hellingwerf, K. J., and Konings, W. N. (1982) *Biochim. Biophys. Acta* **681,** 85–94
Machleidt, H., Roth, S., and Seeman, P. (1972) *Biochim. Biophys. Acta* **255,** 178–189
Overton, E. (1896) *Z. Phys. Chem.* **22,** 189–209
Rekker, R. F. (1977) in *The Hydrophobic Fragmental Constant* (Nauta, W. T., and Rekker, R. F., eds), Elsevier, Amsterdam, The Netherlands
Rekker, R. F., and de Kort, H. M. (1979) *Eur. J. Med. Chem.* **14,** 479–488
Riddick, J. A., Bunger, W. B., and Sakano, T. K. (1986) in *Techniques of Chemistry* (Weissberger, A., ed) Vol. 2, John Wiley & Sons, Inc., New York
Rottenberg, H. (1990) *Biochim. Biophys. Acta* **1018,** 1–17
Rouser, G., Fleischer, S., and Yamamoto, A. (1970) *Lipids* **5,** 494–496
Seeman, P. (1972) *Pharmacol. Rev.* **24,** 583–655
Seeman, P., Roth, S., and Schneider, H. (1971) *Biochim. Biophys. Acta* **225,** 171–184
Shinitzky, M., and Barenholz, Y. (1978) *Biochim. Biophys. Acta* **515,** 367–394
Sikkema, J., and de Bont, J. A. M. (1991) *Biodegradation* **2,** 15–23
Sikkema, J., Poolman, B., Konings, W. N., and de Bont, J. A. M. (1992) *J. Bacteriol.* **174,** 2986–2992
Smith, M. R. (1993) in *Biochemistry of Microbial Degradation* (Ratledge, C., ed) pp. 347–378, Kluwer Academic Press, Dordrecht, The Netherlands
Tomlinson, E., and Hafkenscheid, T. L. (1986) in *Partition Coefficient, Determination and Estimation* (Dunn, W. J., III, Block, J. H., and Pearlman, R. S., eds) pp. 101–141, Pergamon Press, Inc., New York
Uribe, S., Ramirez, J., and Peña, A. (1985) *J. Bacteriol.* **161,** 1195–1200
Uribe, S., Rangel, P., Espínola, G., and Aguirre, G. (1990) *Appl. Environ. Microbiol.* **56,** 2114–2119
Verschueren, K. (1983) *Handbook of Environmental Data on Organic Chemicals*, 2nd Ed., Van Nostrand Reinhold, New York
Yeagle, P. L. (1989) *FASEB J.* **3,** 1833–1842